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Macroinvertebrate Biomonitoring Protocol for Four Prairie Streams

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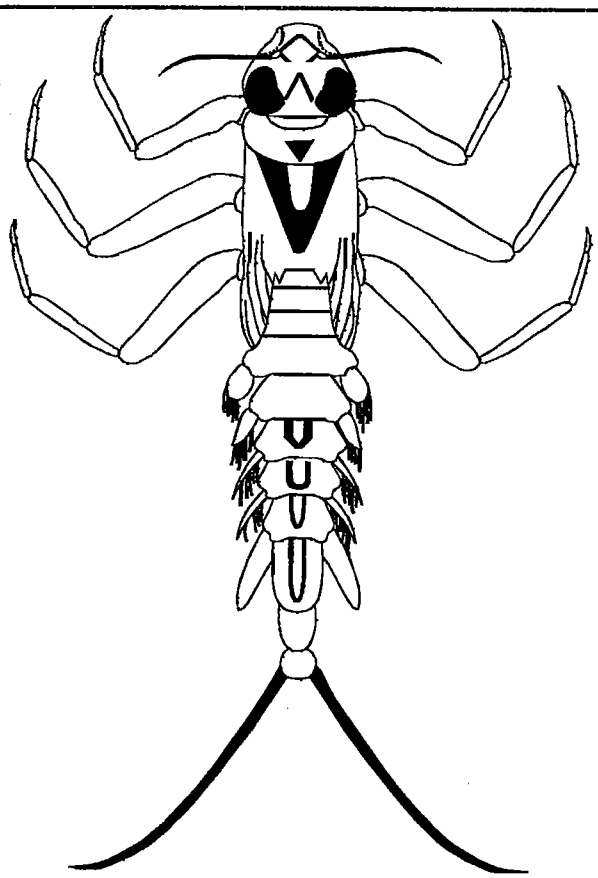

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Macroinvertebrate Biomonitoring Protocol for Four Prairie Streams

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PREFACE

The original draft of the Macroinvertebrate Biomonitoring Protocol for Four Prairie Streams was authored by Dr. James T. Peterson, then of the Missouri Field Station of the Northern Prairie Wildlife Research Center, Biological Resources Division, U.S. Geological Survey, in March 1997. Following USGS/Biological Rources Division scientific peer review and National Park Service management review, the final document was completed by the other authors.

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1.0 INTRODUCTION

1.1 Background

This protocol was developed for use by four of the six national park units comprising the Great Plains Prairie Cluster Long-Term Ecological Monitoring Program (hereafter referred to as Prairie Cluster Program). Three of these parks, Agate Fossil Beds National Monument, Nebraska (AGFO), Homestead National Monument of America, Nebraska (HOME), and Pipestone National Monument, Minnesota (PIPE), have stream resources which are prairie in nature. The fourth Park, Wilson's Creek National Battlefield, Missouri (WICR), has Ozark woodland streams. Prairie streams differ from woodland streams primarily in that shading is generally less in upstream reaches, the reverse of woodland streams, resulting in higher in situ production in upstream areas; the detrital inputs to prairie streams are primarily from grasses rather than leaf litter; and their hydrologic cycles typically have greater extremes in wet/dry conditions (Matthews 1988). Readers desiring more information on the ecology of prairie and Ozark streams are referred to Matthews (1988), Rabeni (1996), and Rabeni et al. (1997).

Although the character of the streams of the Prairie Cluster parks may differ, they are similar in that their watersheds encompass areas outside the park boundaries, subjecting these resources to threats from local land-use practices. The threats facing these streams are numerous (Matthews 1988; Rabeni 1996), and include:

1. Altered flow regimes from changes in land-use which result in increased runoff and sedimentation.
2. Increased light levels along with nutrient loads resulting in greater in situ primary production.
3. Physical structure altered by channelization, levee construction, and loss of wetlands and riparian vegetation.
4. Introduced species which compete with native species.
5. Inputs of organic wastes and synthetic organic contaminants.

This biomonitoring protocol was developed to detect changes in the stream macroinvertebrate community, which may result from one or more of these potential threats.

Biomonitoring uses living organisms as a reflection of the quality of an aquatic environment. It is based on the fundamental assumption that a direct relationship exists between the physical and chemical characteristics of an aquatic ecosystem and the structure of its biotic community (Rosenberg and Resh 1993). For example, organisms in an ecosystem must be able to obtain the available resources to complete the activities necessary for growth, survival, and reproduction. If environmental conditions are unfavorable, or necessary resources are unavailable for a given species, that species may be excluded or eliminated from the ecosystem. Aquatic community structure can be used to (1) assess environmental quality by comparing it to the structure of communities in reference systems, or (2) monitor the quality of an aquatic environment over time by comparing community structure from year to year or to a baseline year.

Many different types of aquatic organisms have been used in biomonitoring (Hellawell 1986). However, aquatic insects are among the most widely used because they can be sampled relatively efficiently and effectively (Resh and McElray 1993); they are widespread in aquatic

environments (Merritt and Cummins 1996); there are a large number of species that have a wide range of responses to environmental impacts (Rosenberg and Resh 1993); and since they are relatively sedentary, they can be used to determine the spatial extent of impacts (Resh and Rosenberg 1989). In addition, since macroinvertebrates are relatively long-lived, the community response to aquatic environmental quality integrates the high variability associated with traditional physical and chemical analyses (Rosenberg and Resh 1996).

1.2 Previous macroinvertebrate biomonitoring

Kolkwitz and Marsson (1908) first used macroinvertebrates to determine the degree of water pollution by organic matter in German waters. In North America, Richardson (1928) first related the community structure of macroinvertebrates in the Illinois River to organic pollution from the cattle industry in Chicago. Both studies described zones of degradation and biotic recovery that contained characteristic fauna. Since these early works, considerable effort has been focused on the development of macroinvertebrate-based indicators that use taxa-specific stress tolerance values weighted by taxa abundance. In Britain, the Trent Index (Woodiwiss 1964) was developed to monitor stream water quality and was modified by Chutter (1972), Anderson et al. (1984), and Hilsenhoff (1988) to incorporate region-specific taxa. Biotic indices have also been developed to measure the effects of specific impacts such as heavy metal (Winner et al. 1980) or pulp mill (Hendricks 1974) pollution. For a thorough review of the history and development of macroinvertebrate biomonitoring, see Rosenberg and Resh (1993) or Harris et al. (1991).

1.3 Objectives of macroinvertebrate biomonitoring

The three objectives of the Prairie Cluster macroinvertebrate biomonitoring protocol are to provide park managers with the data necessary to: (1) determine the annual status of the stream macroinvertebrate community structure to estimate stream water quality, (2) compare current estimates with those of other years, and (3) make periodic summaries and interpretive reports on the long-term trends as monitoring is carried out over time.

2.0 MACROINVERTEBRATE PROTOCOL DESIGN

2.1 Development of protocol

In 1988 and 1989, the National Park Service (NPS) began an intensive program to monitor water quality and macroinvertebrate community structure in prairie streams through a collaborative effort between the NPS Water Resources Division and Colorado State University (Harris et al. 1991). Harris et al. (1991) established the sampling sites and methodology. Sites were chosen at WICR, PIPE, AGFO and HOME. To maintain continuity in their monitoring program, the NPS has continued to use the sites and methodology established by Harris et al. (1991). The collection periods and replication recommended in the protocol were established after extensive statistical analyses of the 1988-1989 data to determine the main sources of variation in macroinvertebrate community structure, to develop the most cost-efficient sampling strategy, and to develop biological criteria for detecting changes in community structure (Peterson 1996). Peterson selected 1989 as the baseline year because it was the earliest year with a reasonably thorough sampling at most parks. Peterson concluded that macroinvertebrate

community structure could be adequately summarized by four biotic indices. These indices were chosen because they represented independent estimates of change with minimal redundancy in the community aspects estimated by each metric, i.e. metrics were avoided if they appeared equally sensitive to the same environmental factors. Seasonal variability was found to be much greater than intra-seasonal variability, so that concentrating multiple samples within a season was recommended. Finally, replicate variability (5 subsamples) per sampling day was found to be much lower than intra-seasonal variability. Thus, the sampling strategy recommended to maximize the collection of information while minimizing cost was to collect 5 replicate samples at each site on each of 3 dates, 30 days apart, during the summer season (Peterson 1996). The Biological Resources Division (BRD) recommends maintaining sampling at the same sites and with basically the same field and laboratory methodology (see methods below) established by Harris et al. (1991), and keeping the sampling intensity and timing recommended by Peterson (1996). With the exception of AGFO, which has a poor 1989 sample, the BRD also recommends retaining 1989 as the baseline year for gauging changes in macroinvertebrate community structure. Although the 1989 baseline does not meet protocol requirements (inadequate numbers of sampling dates and replicates), statistically significant changes in the macroinvertebrate communities can be shown because the changes have been so great (Rizzo 1997a; 1997b, 1997c). Detection of more subtle changes from future sampling efforts will require adherence to the recommendations of the protocol.

2.2 Monitoring sites

Harris et al. (1991) established the macroinvertebrate monitoring sites for the four Prairie Cluster park streams (Table 1). No criteria were given for selection of the actual sites. However, it appears that the sites were chosen to estimate water quality conditions at points of entry and exit of park boundaries, except for the site in Skegg's Branch at WICR which was chosen to assess the influence of a major tributary. Accessibility was probably also a consideration in site selections. A third site at AGFO was established in 1997 to collect data from a stream section which will likely be impacted by highway construction in the future. Continued use of these established sites provides a long-term, site specific sampling record for each park.

To ensure sampling at the same location, each site is permanently marked and the precise location determined using global positioning technology. A photographic record of the site is maintained at each park. Site markers are located at the upstream boundary of each site and consist of a PVC pipe (1 m long, 5 cm diameter) attached with a chain link to a rebar (1 m long, 1.3 cm diameter). Two-thirds of the length of the rebar is driven into the ground, approximately 3 m from the streambank, while the site marker is driven about 30 cm into the ground (Figure 1). The PVC pipe is marked with the park name and site code.

2.3 Choice of sampling device

Choosing the appropriate sampling device is one of the most critical aspects of biomonitoring (Resh and McElray 1993). However, the objectives of this protocol also require continuity with the methods of Harris et al. (1991) for comparison to the 1989 baseline data, and to other past NPS sampling. Park managers wishing to initiate a biomonitoring program could

also use other field methodologies if readily available, e.g. kick nets or Hess samplers, as discussed in Hauer and Lamberti (1996).

The physical characteristics of the stream determine which device is most appropriate. Surber samplers (0.0929 m^2) are used to sample riffles in streams dominated by gravel/rocky substrate and numerous shallow riffle bedforms because riffles are typically more accessible and contain higher biotic diversity. Harris et al. (1991) used Surber nets of differing mesh sizes. At PIPE the capture net mesh size was 1050 µm while 200 µm was used at WICR. Mesh sizes greater than 500 µm risk losing many small macroinvertebrates, particularly the abundant chironomids (Voshell et al. 1989; Hauer and Lamberti 1996); so use of a 200 µm net is recommended. Hester-Dendy samplers are placed in pools or slowly-moving waters (i.e. no riffles) to simulate stream habitats dominated by abundant woody debris. Water depths must be at least 25 cm. These samplers were chosen for use at AGFO and HOME. They are composed of nine 57.76 cm^2 square hardboard plates separated by 3 mm spacers and connected by a long eye-bolt, providing 0.0929 m^2 of surface area for macroinvertebrate colonization. Figure 2 gives a simplified illustration of each sampling device, both of which are available from scientific supply companies.

2.4 Sampling frequency and timing

Three Surber or Hester-Dendy samples, with five replicates per sample, are collected at 30-day intervals during the summer (Peterson 1996). The summer period and spacing of samples was chosen to reflect the period of most rapid benthic macroinvertebrate growth and development. It is controlled by the number of growing degree days (i.e., the number of days the average daily temperature is above a certain threshold, usually 10 °C) and other factors. To maintain sampling consistency with regard to temperature, the summer period at each park is based upon the normal average daily temperatures for the nearest National Weather Service station (Table 2). Sampling periods are scheduled at the beginning of the calendar year.

The initial sample (Surber) or deployment (Hester-Dendy) should be scheduled within about 10 days of the beginning of the sampling window. Subsequent samples are then scheduled 30 days apart. If conditions such as high or low flows prevent a sample collection on the scheduled date, the samples are collected as soon as possible after the scheduled date.

2.5 Habitat assessment and ancillary data

Data collection sheets for each site include: date, park name, site code, and the name(s) of personnel taking part in the sampling (See examples in Appendices A and B). Prior to sampling, the stream gauge-height to the nearest 0.1 m and stream water temperature to the nearest 0.5 °C are recorded. Gauges are purchased from commercial scientific supply vendors or made using a 6-8 ft long 2 x 4 painted white with visible markings every 1 cm. To prevent floodwater damage, gauges are mounted to stable, sturdy structures, such as bridge pillars in the stream channel (Figure 3). Since the gauges in most parks cannot be adequately tied in to benchmarks or quantitatively gauged stream segments, the gauges are used to provide a qualitative estimate of flow conditions based on height readings relative to readings during normal flow conditions, as determined from personal observations. Gauges are mounted during normal flow conditions; thus the gauge readings will be arbitrary.

Prior to deploying the Hester-Dendy sampler or *immediately following* Surber sampling, the investigator conducts a physical habitat evaluation for the stream area adjacent to each sampling site. In addition, a fourth assessment is carried out for Hester-Dendy sites at the time of the final sampler retrieval. The equipment needed for the habitat evaluation includes a data sheet and a standard meter stick (Appendices B and C). The results of readings taken at three randomly selected points within a 0.5 m radius of the sampling location (Figure 4) are recorded and mean depth and current velocity are calculated after the field work is completed. Depth is recorded in the sampling area (Surber) or sampler deployment area (Hester-Dendy) to the nearest 0.5 cm using the meter stick. At the same time, current velocity is estimated by first measuring the vertical displacement of water in the front (upstream side) of the meter stick to the nearest 0.5 cm (Figure 4). The meter stick should be held straight up with the marked side facing the current, by personnel standing downstream of the meter stick. Current velocity is calculated as:

$$C = \ln(V)0.304 + 0.405$$

where C is current velocity (m/s) and ln(V) is the natural log of the average vertical water displacement in mm (Schlosser 1982). Finally, the investigator determines the substrate types and estimates the percent of surface cover within a 1 m radius of the sample site (Figure 5). Substrates are categorized as silt (< 0.06 mm), sand (0.06-2 mm), gravel (2-64 mm), cobble (64-256 mm), or boulder (> 256 mm); (Figure 5; Wentworth 1922).

2.6 Procedure for collecting macroinvertebrates

Sampling procedures were selected to maintain consistency with the procedures used in previous park sampling (Boyle et al. 1990). This biomonitoring protocol uses several metrics to characterize invertebrate communities because metrics differ in their sensitivity to changes in different environmental variables. For example, some metrics may be more sensitive to changes in structural variables such as sediment grain size, than to chemical and physical water quality variables (Bode and Novak 1995; Yoder and Rankin 1995).

2.6.1 *Surber sampler-* In riffles, benthic macroinvertebrate samples are collected with the Surber sampler. Riffles are shallow areas of a stream with relatively fast current and gravel/rocky substrate. Since macroinvertebrates are mobile their abundance at a site is subject to change from disturbance. To prevent disturbing a site, the area upstream of the sampling site is avoided prior to sampling. For this reason, the habitat assessment is carried out *after* Surber sampling. The investigator begins at the fifth riffle downstream from the site marker and moves upstream, collecting one sample per riffle until five samples have been collected. Riffles chosen for sampling should be at least 1 m apart to avoid disturbing a new site while sampling downstream riffles, and to avoid pseudoreplication where a contiguous riffle is sampled as an additional "separate" site.

The equipment needed for Surber sampling includes a vegetable brush, a small hand rake, forceps, a wash bottle, and five large, pre-labeled wide-mouth jars (e.g., 500-1000 ml jars made of inert plastic; Appendix C). Labels are affixed to the jars and an identical label is placed inside each jar. Labels indicate the park code, monitoring site code, date, replicate number, and type of sample fixative (Appendix D). Waterproof paper (available from scientific supply companies) and a #2 lead pencil are used for the labels. The sampler is placed in areas of the riffle with sufficient depth and current to wash dislodged organisms into the catch net, but not so deep that

the sampler is submerged. The sampler should seal off the streambed so that no organisms escape between the bottom of the sampler and the streambed. With the sampler in place, each individual rock (i.e. cobble and larger particles) is scrubbed underwater with the vegetable brush to dislodge organisms and wash them into the catch net. Each rock is then inspected and any remaining organisms are removed with the forceps. After all the rocks are brushed and inspected, the streambed within the sampler is disturbed with the hand rake to a depth of 5 cm. The contents of the catch net are washed to the end of the net by splashing water along the sides of the net or by briefly submerging the net. The net contents are placed into the wide-mouth jar by inverting the catch net. To get all of the contents into the jar, it is necessary to rinse the net with the squeeze bottle. Finally, the catch net is inspected for any remaining organisms which are removed with the forceps and placed in the jar. Prior to leaving each site, samples and data sheets are rechecked to ensure that they have been properly labeled and filled out, respectively.

2.6.2 *Hester-Dendy sampler-* The equipment needed for the Hester-Dendy sampling includes a nylon rope, a knife, a wash bottle, a U.S. standard sieve with 212 μm mesh, forceps, and five large, pre-labeled wide-mouth jars (Appendix C). Labeling follows the procedures described above for Surber sampling. The Hester-Dendy samplers require a deployment and retrieval trip to complete one invertebrate sample. Because of the 30-day colonization period, Hester-Dendy samplers for the second and third sampling periods are deployed when retrieving the samplers from the first and second sampling periods, respectively. Each of the five Hester-Dendy samplers is deployed separately in the streams at sites with water depths greater than 25 cm and with slow to moderate current. The samplers should be spaced at least a meter apart. The samplers are deployed using the nylon rope, tied to the eye-bolt, and suspended from sturdy woody vegetation or other support, such that they are submerged at least 6 cm above the streambed, and 6 cm below the surface. The samplers are placed while working upstream. To avoid disturbing a site, the site is avoided prior to sampler deployment.

After the colonization period, the samplers are retrieved by placing the dipnet slightly below and downstream of the sampler. The sampler and dipnet are carefully lifted out of the water and placed into the large wide-mouth jar. After removal from the jar, the dipnet is inspected for any remaining organisms which are removed with the forceps and placed in the jar. Prior to leaving each site, samples and data sheets are rechecked to ensure that they have been properly labeled and filled out, respectively.

2.7 Sample preparation and shipping

Sample fixation is now done using ethanol rather than formalin because use of formalin would expose personnel to a potent carcinogen. When not in use, ethanol is required to be stored in a flammable-proof metal storage cabinet. Ethanol-fixed samples must be rehydrated 48-72 hours after the sample is collected. Rehydration consists of decanting the original 80% ethanol and refilling the sample jar with tap water. Following a 0.5-1 h rehydration period, the water is decanted and the sample jar refilled with new 80% ethanol solution. Decant the sample by pouring the contents through a U.S. Standard sieve with 212 μm mesh, and washing the sieve contents back into the jar.

As soon as possible the macroinvertebrate samples are placed in airtight containers and shipped to the macroinvertebrate identification contractor along with copies of the habitat data

sheets. Since the samples contain ethanol, they are subject to hazardous material regulations. Samples shipped by ground transportation fall under the purview of 49CFR173.4. These regulations contain an important provision for shipment of samples with small volumes of preservative (or other regulated materials). An unlimited number of samples containing 30-ml or less of preservative may be packed and shipped by personnel with no formal training in hazardous material packing. Ground transportation (UPS ground; U.S. Mail) must be specified when the shipper is contacted. Samples should be shipped in HDPE plastic bottles recommended by the manufacturer as suitable for shipping. Lids should be screwed on tightly, and absorbent packing material should enclose the bottles. Carriers using air transportation fall under a different set of regulations entirely, and this method of shipping is not recommended. In addition, shipping samples with preservative volumes greater than 30-ml require completion of a formal training course in packing hazardous materials. A record of all shipped samples should be logged by the park resource manager. Copies of the sample log and the habitat data sheets should be sent to the identification contractor along with the samples. An additional copy of the habitat data sheets is sent to the Prairie Cluster Program office (Table 3). The park resource manager keeps the original habitat data sheets and shipping information on file (Table 3).

3.0 LABORATORY SAMPLE PROCESSING OF MACROINVERTEBRATES

3.1 Sample cleaning and sorting

Sample processing is the responsibility of the macroinvertebrate identification contractor. The contents of each sample jar (i.e., invertebrates, organic debris, Hester-Dendy sampler) are poured into a U.S. standard sieves (U.S. standard No. 60, 212 μm mesh for Hester-Dendy samples; for Surber samples mesh sizes equal to or smaller than those of the Surber capture net are used). At this point, the Hester-Dendy sampler is unscrewed and the hardboard plates removed from the eye-bolt. Both sides of the plates are rinsed over the sieve and the plates carefully inspected for any remaining organisms. The sample contents in the sieve are rinsed into an appropriate container with clean water. If the sample is not sorted immediately, it is stored in a properly labeled container filled with 80% ethanol. The internal label is kept with the sample at all times. Duplicate waterproof labels will be printed in advance and supplied to the contractor by the Prairie Cluster Program staff.

Each sample is picked and sorted individually to avoid mixing the contents of different samples. A portion of each sample is transferred to a clear petri dish or modified zooplankton wheel and carefully examined under a dissecting microscope with at least 20X magnification. The macroinvertebrates are separated from the debris and placed into vials containing 80% ethanol. This process is repeated until the entire sample has been examined. Each vial contains a waterproof label with park code, monitoring site code, date, and replicate number.

In some samples, large sample volumes and/or high densities of some macroinvertebrate taxa, particularly chironomids, necessitate subsampling (when abundance of a taxonomic group greatly exceeds 100 individuals) to prevent unacceptable processing backlogs. The subsampling procedure, including precision and accuracy estimates are those described in Plafkin et al. (1989). Briefly, subsampling is undertaken by first distributing the entire sample uniformly within a suitable sized container, and removing any large pieces of debris. Next, a grid composed of ten

equally-sized cells is placed over the sample container. Grid cells are then randomly selected for processing (e.g. roll of a ten-sided die; random number table, etc). Cells are processed until cursory observation shows that about 100 organisms (± 20%) have been collected. In all cases, the percentage of the total sample removed for processing must be recorded.

3.2 Macroinvertebrate identification

After sorting, an identifier with formal training in aquatic macroinvertebrate taxonomy identifies the macroinvertebrates. A dissecting microscope and taxonomic keys are used to identify each specimen to the taxonomic level shown in Table 4. In most cases the entire sample can be identified to the required level, however, an occasional sample may contain early instars or damaged specimens which cannot be identified to the required level (C. F. Rabeni, personal communication). In those cases where complete identification is impossible, identifications should be carried out to the lowest possible level. Most insect specimens are identified to genus level. Most aquatic and semiaquatic insect orders are identified using the keys in Merritt and Cummins (1996), whereas non-insect macroinvertebrates are identified using Pennak (1989). Additional taxonomic references that are used to identify specimens in specific orders include Edmunds et al. (1976) for ephemeroptera , Stewart and Stark (1988) for plecoptera, and Wiggins (1977) for trichoptera. This taxonomic level is similar to that used by Harris et al. (1990), except that ephemeroptera and plecoptera are identified only to genus instead of to species, and diptera are identified only to family. Rabeni et al. (1997) found that greater taxonomic resolution of dipterans had no impact on biotic indices, and thus did not justify the greater cost and effort. However, chironomids should be tabulated by blood group (red-blooded chironomids and all other chironomids), because the two groups have different pollution tolerance scores (Hilsenhoff 1988). During the identification process, a running total of each taxon in each replicate sample is recorded on a data sheet that is signed or initialed by the identifier. The identification contractor will also enter the data into an ACCESS database. The database structure and screen forms for data entry will be provided by the Prairie Cluster Program data manager. The contractor will provide a list of any new taxa entered into the database along with their new taxa codes. A current list of taxa in the database, along with their respective codes, is shown in Appendix E. Representative specimens for each park, verified by a taxonomic specialist, are preserved in a reference collection to ensure accurate identifications.

3.3 Sample transfer and storage

Following identification, each sample is separated by taxon and stored in vials. The samples in each vial are preserved in 80% ethanol and each resulting vial contains a duplicate copy of the complete collection label. Also, the original internal label is kept with the vials containing the corresponding sample. The sorted and identified samples, sample log, macroinvertebrate reference collection, original paper data sheets, and ACCESS database are sent to the Prairie Cluster Program office. The Hester-Dendy samplers, sample bottles, copies of the data sheets, and a copy of the sample log are returned to each park. The contractor keeps a copy of the sample log, the data sheets, and a disk copy of the database. The reference collections will be housed in a permanent collection at the Prairie Cluster Program office for at least 5 years.

3.4 Deadlines

Data sheets with the total number of specimens by taxon and replicate are sent to the Prairie Cluster Program office within 4 months of receiving the samples from the last sampling period (Table 3). The macroinvertebrate identification contractor keeps copies of the macroinvertebrate data sheets and shipping information on file (Table 3).

4.0 CRITERIA IMPLEMENTATION

4.1 Biological criteria indices

This biomonitoring protocol uses several metrics to characterize macroinvertebrate communities because metrics differ in their sensitivity to changes in different environmental variables. For example, some metrics may be more sensitive to changes in structural variables such as sediment grain size, than to chemical and physical water quality variables (Bode and Novak 1995; Yoder and Rankin 1995).

4.1.1 *Total density* - Total density of macroinvertebrates across taxa has been used to assess stream quality (Plafkin et al. 1989). It is calculated only for samples collected with the Hester-Dendy sampler. In general, macroinvertebrate densities decrease when communities are exposed to a stress such as water pollution or habitat alteration (Resh and Grodhaus 1983). Total density of macroinvertebrates across taxa is calculated for each replicate sample by dividing the total number of macroinvertebrates collected by the total area of each Hester-Dendy sampler, i.e. by 0.0929 m^2.

4.1.2 *Biotic Indices* - Biotic indices are commonly used as indicators of water quality (Resh and Jackson 1993; Resh and McElray 1993). In fact, Jones et al. (1981), who simultaneously measured macroinvertebrate community structure and water quality variables in Missouri Ozark streams, found the biotic indices to be more sensitive and less variable than diversity indices for discriminating differences in stream water quality. The family biotic index (FBI) uses taxa specific (e.g., family, genus) pollution tolerance values (Hilsenhoff 1988; Appendix F) to calculate index scores, which are then related to stream water quality (Table 5). The FBI score for each replicate sample is calculated as:

$$FBI = (\Sigma n_i a_i / N) - 0.18$$

where N is the total number of individuals in the sample, n_i is the total number of individuals and a_i the tolerance value for the ith family.

4.1.3 *EPT ratio* - The ratio of Ephemeroptera, Plecoptera, and Trichoptera (EPT) abundance (numbers of individuals) to Chironomidae abundance has also been used as a stream water quality indicator (Resh and Grodhaus 1983). It is calculated only for replicate Surber samples. In general, EPT taxa are relatively pollution intolerant, whereas Chironomidae are pollution tolerant. Thus, higher values indicate better stream water quality. The EPT ratio (R) is calculated for each replicate Surber sample as:

$$R = EPT/[EPT + C]$$

where EPT is the abundance of EPT taxa, and C is the abundance of Chironomidae.

4.1.4 *Taxa richness* - Richness can be a useful criterion to describe the biological quality of a stream (Resh and Grodhaus 1983). Low richness may indicate that a stream has been subjected to one or more stresses. Taxa richness is simply the sum of the number of families

represented in a Surber sample replicate, or the number of genera represented in a Hester-Dendy sample replicate.

4.1.5 *Taxa diversity* - Diversity is a measure of how the total number of individuals in a sample are distributed among the total species in the sample. Maximum diversity occurs in a community when the number of individuals is distributed as evenly as possible among species (Pielou 1966). High diversity indicates better stream quality (Resh and Jackson 1993). The Shannon-Wiener diversity index (H′) is calculated for each replicate sample as:

$$H' = -\Sigma(n_i/N)\ln(n_i/N)$$

where N is the total number of individuals in the sample, and n_i is the total number of individuals in the *i*th family or genus for samples collected with the Surber and Hester-Dendy samplers, respectively.

4.2 Evaluation of annual changes in stream water quality

The changes in the macroinvertebrate community of each stream are evaluated using analysis of variance (ANOVA). Means based on the fifteen data point calculations for each metric of the current-year data are tested against the appropriate baseline means (i.e. 1989 or other baseline year). Data from other years may be included for comparison if desired. The selected data set is tested for the assumptions of parametric analyses prior to carrying out the ANOVA, and the data is transformed, if required. If multiple years of data are tested and found significant, paired data comparisons can be carried out using t-tests, provided that the probability level is adjusted for the number of comparisons made. Analysis of multiple years allows assessment of the status of the current-year data to that of past years in addition to the baseline year. Since different indices may reflect different aspects of environmental change, at least two indices must be significantly ($P \leq 0.05$) different from the baseline before a conclusion of significant impact or improvement to the community is reached.

4.3 Determination of impact source

When significant differences from the baseline data in two or more community metrics is found by the statistical testing, the physical habitat data and streamflow data from U.S. Geological Survey gauging stations (Table 6) are reviewed to assess whether natural events may be responsible for the changes. For example, naturally occurring events such as severe floods or droughts can negatively affect macroinvertebrate communities (Resh et al. 1988). If the statistical findings cannot be resolved by analysis of the physical habitat and streamflow data, a research project may be necessary to determine the cause.

Also, it is possible that different metrics can give conflicting results. An increase in density or richness may not indicate an improvement in water quality if the increase is due to larger numbers of pollution tolerant chironomids for instance. Since the protocol is designed primarily as a tool for detection of unfavorable impacts, a significant increase in a particular metric would not be interpreted as an improvement if two other metrics indicate deteriorated water quality.

4.4 Annual report

By March 1 of each year following the sampling period, the Prairie Cluster Program staff will provide park managers with an annual report on the current status of the biological criteria indices vs. the baseline. The reports will also evaluate the habitat data for any potential environmental sources of impact should significant changes be detected. An example of such a report is given in Appendix H.

5.0 INTEGRATION WITH OTHER STUDIES AND MONITORING

5.1 Related monitoring

Daily weather information is currently collected by automated weather stations at AGFO, PIPE, and WICR. In the future, weather data and other environmental data from external sources (USGS streamflow data) will provide the high quality data that are critical to elucidate potential impact sources. For example, with time, streamflow data can be related to gauge readings providing a method of assessing flow conditions continuously, not just on sampling dates.

5.2 Changes to biomonitoring protocol

Changes in biological indices reflect statistically significant changes in macroinvertebrate community structure that indicate changes in stream water quality. However, biological significance is only inferred from monitoring data and must be assessed by comparing the variability of indices among streams with similar water quality (e.g., good, fair, poor) to baseline data from a stream with demonstrably good water quality. Since most of the Prairie Cluster parks are located in states in which region-specific biological criteria and reference stream conditions are being established (Southerland and Stribbing 1995), Prairie Cluster Program personnel may wish to integrate this information into the monitoring protocol in the future. Also, since stream water quality could also improve through time, the Prairie Cluster Program staff may wish to reevaluate the baseline dataset and sample size requirements periodically.

5.3 Quality assurance

Consistency is one of the most important aspects of stream monitoring protocols. Differences in gear operation, specimen identification, and data entry can lower data quality by increasing variance (Hannaford and Resh 1995). To ensure high quality data, macroinvertebrate sampling and habitat evaluation training sessions are held every 3-5 years, or when a new resource manager arrives at a park.

In addition, long-term trend analyses require that data be archived in a standard format at one location accessible to all the parks. The Prairie Cluster Program office uses an ACCESS database structured as shown in Table 7. Other statistical or spreadsheet programs can also be used. However, it is important for continuity or "institutional memory" that metadata descriptions be incorporated into the database. The Prairie Cluster ACCESS database is designed for compatability with other NPS Prairie Cluster monitoring databases. The tabular structures of this database include site tables (sampling sites); event tables (sampling dates); a record table which contains the habitat assessment data, and assigns a unique record number to each combination of site, sampling date, and replicate; the density table which contains the data (in numbers per m^2 x

10) by taxoncode and record number; and a taxa table which includes unique codes for each taxon. This table must be updated annually as new taxa are added. . Data entry is accomplished by using customized forms which prevent duplicate or incorrect data entry for all data except density. The Prairie Cluster Program office checks for density transcription errors by cross checking the electronic database with 10% of the original hardcopy datasheets. Data from various tables can be queried to produce a new table suitable for export into the statistical packages required for the data analyses in this protocol. This database also contains built-in metadata descriptions of the dataset through the use of comment/description fields in the appropriate tables.

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Table 1. Names, codes, locations, and sampling methods for macroinvertebrate biomonitoring in four prairie streams, from Harris et al. (1991). Hester-Dendy samplers are used at AGFO and HOME, Surber samplers at PIPE, and WICR.

Park name	Park Code	Site Code	Location
Agate Fossil Beds National Monument	AGFO	AFB1	Niobrara River, 110 m west of Hwy 29 bridge
		AFB3	Niobrara River, 25 m above visitor's trail crossing
Homestead National Monument	HOME	HMS1	Cub Creek, SW corner of park
		HMS2	Cub Creek, 50 m NE of Hwy 4 before exiting park
Pipestone National Monument	PIPE	PPS1	Pipestone Creek, riffle 30 m below Winnewissa Falls near a large willow tree
		PPS2	Pipestone Creek 25 m below Lake Hiawatha directly above Circle Trail crossing
Wilson's Creek National Battlefield	WICR	UPPER	Wilson's Creek, north of first crossing of visitor's road 0.4 km from creek entrance into park
		LOWER	Wilson's Creek, 25 m south of second crossing of visitor's road
		SKEGGS	Skegg's Branch, 10 m from road ZZ bridge

Table 2. Summer sampling period for macroinvertebrate biomonitoring in four prairie streams.

Park name	Summer Period	Growing degree Days [1]	NWS Station [2]
Agate Fossil Beds National Monument	6/18-9/19	301-1378	North Platte, NE
Homestead National Monument	6/18-9/19	302-1376	Omaha, NE
Pipestone National Monument	6/18-9/18	307-1375	Sioux Falls, SD
Wilson's Creek National Battlefield	6/1-8/10	346-1380	Springfield, MO

[1] Growing degree days calculated using normal average daily temperature and threshold of 10 °C.
[2] The nearest National Weather Service stations for which normal average daily temperature data were available.

Table 3. Responsibilities of macroinvertebrate biomonitoring cooperators.

Cooperator Responsibilities

Park Resource Manager

(1) Plan field sampling trips and maintain equipment
(2) Collect benthic macroinvertebrate samples and measure physical habitat
(3) Re-hydrate samples after 48-72 hrs, prior to shipping samples to contractor
(4) Ship samples and data sheets to the identification contractor
(5) Send data sheets to Prairie Cluster LTEM staff
(6) Archive data sheets and picked/sorted samples for a minimum of 1 year
(7) Maintain macroinvertebrate reference collection provided by the identification contractor

Prairie Cluster LTEM Program Staff

(1) Assist resource managers in planning sampling dates prior to the summer sampling season
(2) Check subset of contractor data sheets for errors.
(3) Analyze and interpret macroinvertebrate data
(4) Provide annual reports, periodic summaries, and recommendations to each park
(5) Archive data in a standardized format
(6) Periodically hold macroinvertebrate sampling and habitat evaluation training sessions for resource managers
(7) Periodically reevaluate baseline values and sample size requirements for each park

Identification Contractor (Private/University Laboratory)

(1) Pick, sort, and identify macroinvertebrate samples to the required taxonomic level
(2) Record the number of macroinvertebrates, by replicate and taxon, on data sheets signed by the sorter/identifier
(3) Send invertebrate data sheets with the total number of specimens by taxon and replicate to the Prairie Cluster LTEM Program office within 4 months of receiving last sample
(4) Provide a macroinvertebrate reference collection for each park
(5) Ship picked/sorted samples, stored in 80% ETOH, and invertebrate data sheets back to each park

Table 4. Level of taxonomic resolution used in benthic macroinvertebrate sample processing.

Collembola:	Order
Ephemeroptera:	Genus
Plecoptera:	Genus
Hemiptera:	Genus
Megaloptera:	Genus
Trichoptera:	Genus
Lepidoptera:	Genus
Coleoptera:	Genus
Odonata	Genus
Neuroptera	Genus
Diptera:	Family
Nematoda:	Phylum
Turbellaria:	Genus
Annelida:	Class
Acarina:	Class
Isopoda:	Genus
Amphipoda:	Genus
Decapoda:	Family
Gastropoda:	Family
Pelecypoda:	Family

Table 5. Water quality ratings using the family biotic index (FBI), from Hilsenhoff (1988)

FBI	Water Quality	Degree of Organic Pollution
0.00-3.75	Excellent	Organic pollution unlikely
3.76-4.25	Very good	Possible slight organic pollution
4.26-5.00	Good	Some organic pollution likely
5.01-5.75	Fair	Fairly substantial pollution likely
5.76-6.50	Fairly poor	Substantial pollution likely
6.51-7.25	Poor	Very substantial pollution likely
7.26-10.00	Very poor	Severe organic pollution likely

Table 6. U.S. Geological Survey gauging stations closest to the prairie stream biomonitoring sites.

Park	Stream	Gauge name	Gauge ID#
Agate Fossil Beds National Monument	Niobrara River	Niobrara River at Agate Fossil Beds NM, NE	06454100
Homestead National Monument	Cub Creek	Big Blue River at Beatrice, NE	06881500
Pipestone National Monument	Pipestone Creek	Split Rock Creek at Corson, SD	06482610
Wilson's Creek National Battlefield	Wilson's Creek, Skegg's Branch	Wilson's Creek near Battlefield, MO Wilson's Creek near Battlefield, MO	07052160 07052160

Table 7. An example of an ACCESS database and its tables and linkages for storage and handling of the macroinvertebrate biomonitoring data.

Tbl_Taxa	**Tbl_Density**	**Tbl_Record_ID**	**Tbl_Site**
Taxoncode 1	**Record_ID** ∞	1 **Record_ID**	Site_ID
Order	∞ **Taxoncode**	**Site_ID** ∞	1 **Sitecode**
Family	Density	**Event_ID** ∞	Parkcode
Genus		**Replicate**	Parkname
Species		Temperature	GPS_Latitude
		Current Velocity	GPS_Longitude
		% Cobble	Description
		% Gravel	
		% Sand	**Tbl_Event**
		% Silt	1 Event_ID
		Gauge Height	**Eventcode**
		Depth	Date
		Field Notes	Month
			Day
			Year
			Collector 1
			Collector 2
			Comments

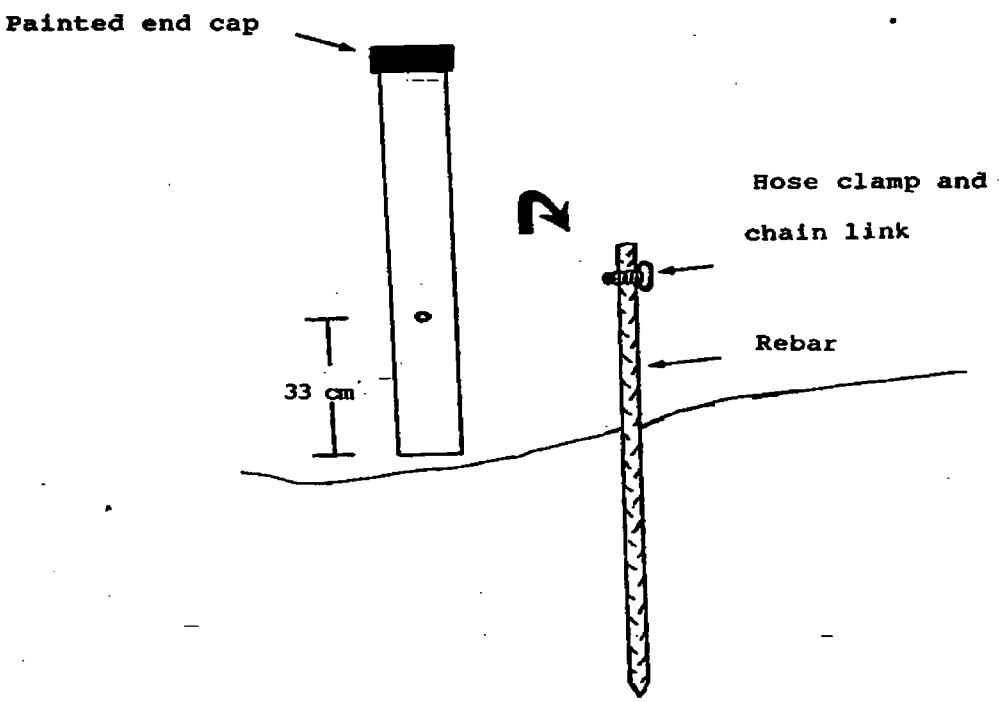


Figure 1. Stream-side marker used to mark stream monitoring sites in Prairie Cluster parks, from Voshell and Hiner (1990).

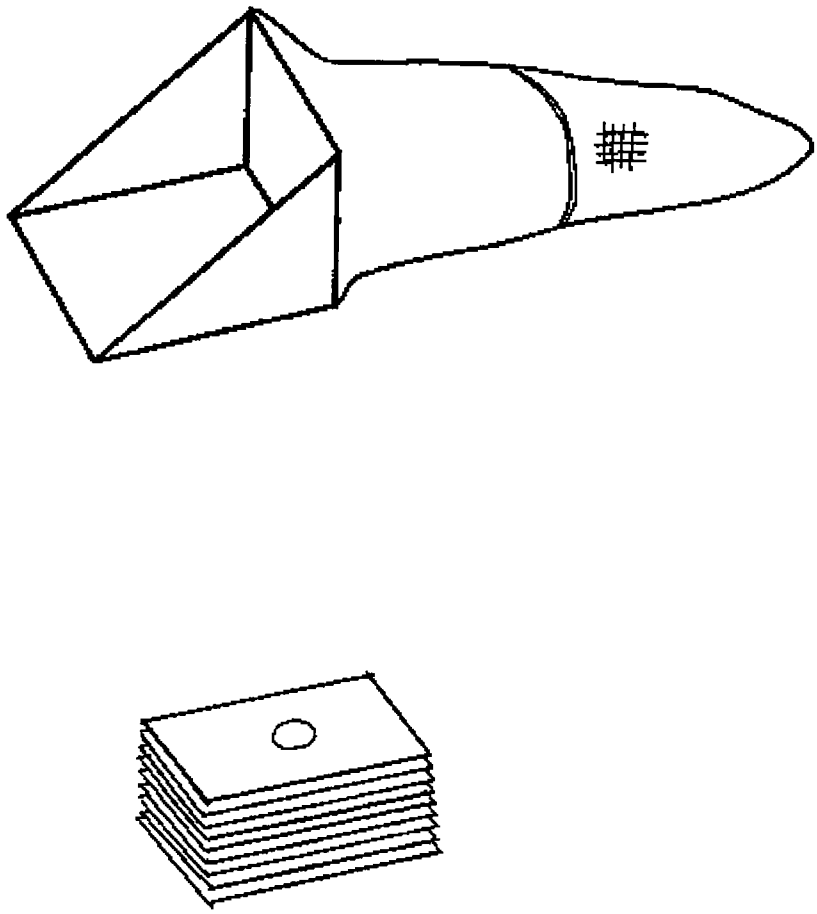


Figure 2. Generalized depiction of a Surber sampler (top), and a Hester-Dendy sampler (bottom).

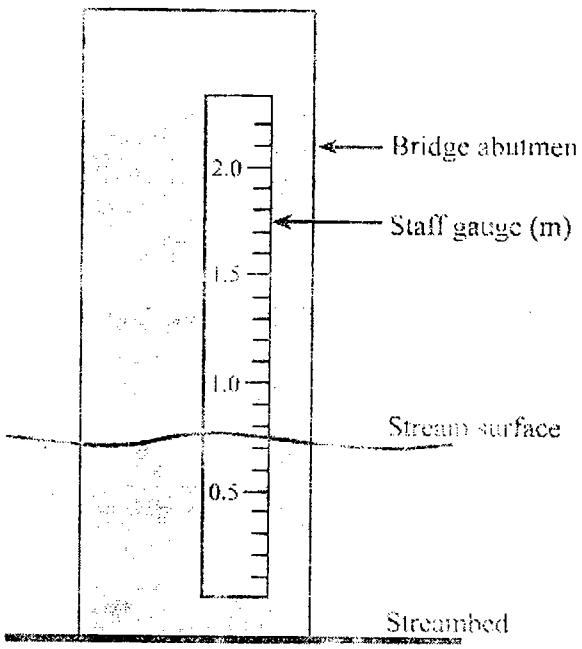
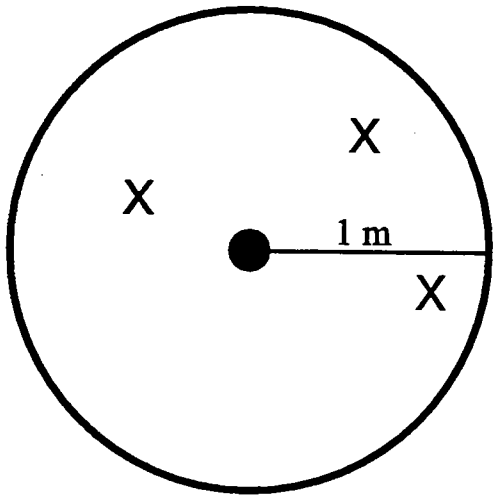


Figure 3. Example of staff gauge placement against a bridge abutment. The initial stream surface, or zero point, is arbitrary.



● Location of Surber sample or Hester-Dendy sampler

X Randomly selected areas for depth and current measurements

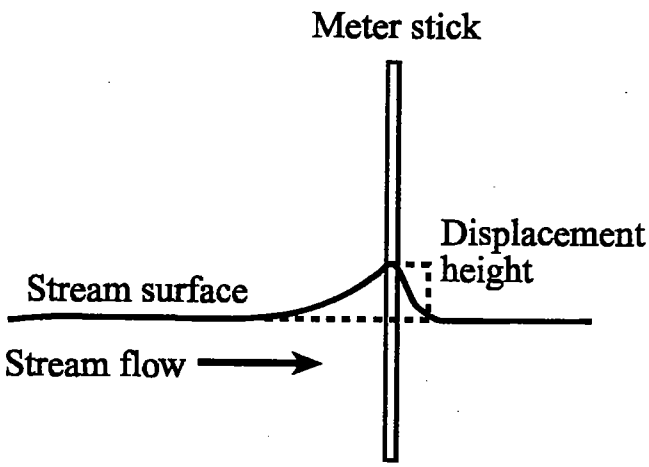
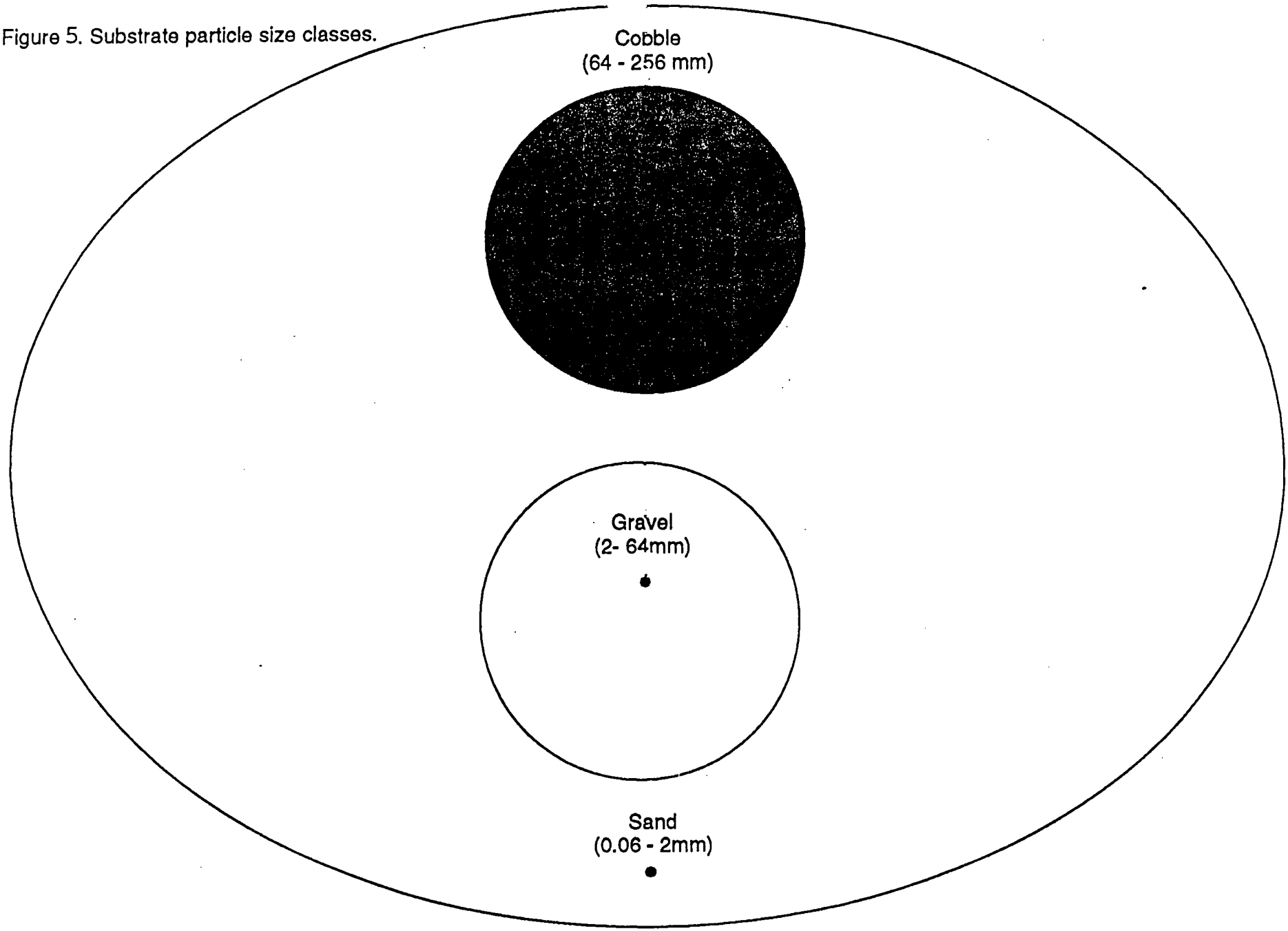


Figure 4. Location of sampling site and habitat assessment sites (top), and illustration of stream velocity measurement (bottom).

Figure 5. Substrate particle size classes.



Appendix A. Surber Sampler Physical Habitat Data Sheet

Park name _____ Monitoring site code _ _____

Date _____ Field crew _____

Water temperature (°C) _____ Gauge height (m)_____

GPS latitude _____ GPS longitude_____
Site description _____
Note: Depth and current estimated within 0.5 m of sampling site, substrate type within 1.0 m of sampling site.

Physical Habitat Data

Replicate #1	Replicate #2	Replicate #3	Replicate #4	Replicate #5
Depth (cm)	Depth (cm)	Depth (cm)	Depth (cm)	Depth (cm)
1 _____	1 _____	1 _____	1 _____	1 _____
2 _____	2 _____	2 _____	2 _____	2 _____
3 _____	3 _____	3 _____	3 _____	3 _____
Avg. _____	Avg. _____	Avg. _____	Avg. _____	Avg. _____
Vertical Displacement (mm)	Vertical Displacement (mm)	Vertical Displacement (mm)	Vertical Displacement (mm)	Vertical Displacement (mm)
1 _____	1 _____	1 _____	1 _____	1 _____
2 _____	2 _____	2 _____	2 _____	2 _____
3 _____	3 _____	3 _____	3 _____	3 _____
Avg. _____	Avg. _____	Avg. _____	Avg. _____	Avg. _____
Substrate (%) Composition	Substrate (%) Composition	Substrate (%) Composition	Substrate (%) Composition	Substrate (%) Composition
Silt _____	Silt _____	Silt _____	Silt _____	Silt _____
Sand _____	Sand _____	Sand _____	Sand _____	Sand _____
Gravel_____	Gravel_____	Gravel_____	Gravel_____	Gravel_____
Cobble_____	Cobble_____	Cobble_____	Cobble_____	Cobble _____

Appendix B. Hester-Dendy Sampler Physical Habitat Data Sheet

Park name _____ Field crew _____

Date _____ Gauge height (m) _____

Monitoring site code _____ Site description _____

GPS latitude _____ GPS longitude_____

Comments_____

_____

Note: Depth and current measurements within 0.5 m of the sampling site.

Substrate types estimated within 1.0 m of the sampling site.

Physical Habitat Data

Water temperature (°C) _____

Depth (cm)	Vertical Displacement (mm)	Substrate Composition (%)
1 _____	1 _____	Silt _____
2 _____	2 _____	Sand _____
3 _____	3 _____	Gravel _____
Avg. _____	Avg. _____	Cobble _____

Appendix C. Field Equipment Check Sheet

General Equipment

Waders or hip boots _____

#2 pencil _____

Habitat Measurement

Habitat data sheets (1 per site) _____

Thermometer _____

Meter Stick _____

Invertebrate Sampling

Surber Sampling

1 wash bottle _____

5 screw-top wide-mouth plastic jars, 750 ml, (1 per replicate) _____

5 pre-printed waterproof labels _____

80% ethanol, ca. 5-L _____

1 forceps _____

1 hand rake _____

1 Surber sampler _____

1 vegetable brush _____

212 µm mesh standard sieve _____

Hester-Dendy Sampling

1 wash bottle _____

5 1-L wide-mouth nalgene jars (1 per replicate) _____

5 pre-printed waterproof labels _____

80% ethanol, ca. 5-L _____

1 forceps _____

1 knife or scissors _____

5 Hester-Dendy samplers _____

5 nylon ropes _____

212 µm mesh standard sieve _____

1 dipnet _____

Appendix D. An Example Label for Inside and Outside of the Macroinvertebrate Sample Container

Park Code: PIPE	**Site Code: PPS1**
Date: 7/4/99	**Replicate #: 1**
Site Description:	**Pipestone Creek - Winnewissa Falls**
Initials of Field Crew:	

Appendix E. Current Taxa Table from the Macroinvertebrate ACCESS Database.

TaxonCode	Order	Family	Genus	Species name
ACAR	Hydracarina	Acarina		
ACAR	Hydracarina	Acarina		
ACARBR	Hydracarina	Acarina	Brachypoda	
ACARHY	Hydracarina	Acarina	Hydrachna	
ACARKA	Hydracarina	Acarina	Kawamuracaras	
ACARLI	Hydracarina	Acarina	Limnochares	
ACARTO	Hydracarina	Acarina	Torrenticola	
ACARTR	Hydracarina	Acarina	Trichothyas	
AESHAEP	Odonata	Aeshnidae	Aeshna	palmata
AESHAN	Odonata	Aeshnidae	Anax	
AESHBO	Odonata	Aeshnidae	Boyeria	
AMELAM	Ephemeroptera	Ameletidae	Ameletus	
AMP	Amphipoda			
AMPHAM	Coleoptera	Amphizoidae	Amphizoa	
ANCY	Gastropoda	Ancylidae		
ANCYFE	Gastropoda	Ancylidae	Ferrissia	
ASELAS	Isopoda	Asellidae	Asellus	.
ASELASO	Isopoda	Asellidae	Asellidae	occidentalis
ASELCA	Isopoda	Asellidae	Caecidotea	
ASELLI	Isopoda	Asellidae	Lirceus	
ASELLIH	Isopoda	Asellidae	Lirceus	hoppinae
ATHEAT	Diptera	Athericidae	Atherix	
ATURAT	Hydracarina	Aturidae	Aturus	
BAET	Ephemeroptera	Baetidae		
BAETAC	Ephemeroptera	Baetidae	Acentrella	
BAETBA	Ephemeroptera	Baetidae	Baetis	
BAETBAB	Ephemeroptera	Baetidae	Baetis	bicaudatus
BAETBAF	Ephemeroptera	Baetidae	Baetis	flavistriga
BAETBAI	Ephemeroptera	Baetidae	Baetis	insignificans
BAETBAR	Ephemeroptera	Baetidae	Baetis	tricorythodes

BAETBAT	Ephemeroptera	Baetidae	Baetis	tricaudatus
BAETDIH	Ephemeroptera	Baetidae	Diphetor	hageni
BAETPA	Ephemeroptera	Baetidae	Paracloedes	
BAETPR	Ephemeroptera	Baetidae	Procleon	
BELOBEF	Hemiptera	Belostomatidae	Belostoma	flumineum
BITHBI	Gastropoda	Bithyniidae	Bithynia	
BRA	Branchiobdellida			CLASS
CAENCA	Ephemeroptera	Caenidae	Caenis	
CAENCAA	Ephemeroptera	Caenidae	Caenis	amica
CALO	Odonata	Calopterygidae		
CALOCA	Odonata	Calopterygidae	Calopteryx	
CALOHE	Odonata	Calopterygidae	Hetaerina	
CAMB	Decapoda	Cambaridae		
CAMBOR	Decapoda	Cambaridae	Orconectes	
CAMBORH	Decapoda	Cambaridae	Orconectes	harrisoni
CAMBORL	Decapoda	Cambaridae	Orconectes	luteus
CAMBORV	Decapoda	Cambaridae	Orconectes	virilis
CERA	Diptera	Ceratopogonidae		
CERAAT	Diptera	Ceratopogonidae	Atrichopogon	
CERABE	Diptera	Ceratopogonidae	Bezzia	
CERACE	Diptera	Ceratopogonidae	Ceratopogon	
CERAMA	Diptera	Ceratopogonidae	Mallochohelea	.
CERAPR	Diptera	Ceratopogonidae	Probezzia	
CHAUNI	Megaloptera	Chauliodinae	Nigronia	
CHIO	Diptera	Chironomidae	Chironomini	
CHIOBE	Diptera	Chironomidae	Beardius	CHIRONOMINI
CHIOCH	Diptera	Chironomidae	Chironomus	CHIRONOMINI
CHIOCR	Diptera	Chironomidae	Crytochironomus	CHIRONOMINI
CHIOCY	Diptera	Chironomidae	Cryptotendipes	CHIROMINI
CHIODI	Diptera	Chironomidae	Dicrotendipes	CHIRONOMINI
CHIOEI	Diptera	Chironomidae	Einfeldia	
CHIOGL	Diptera	Chironomidae	Glypotendipes	CHIRONOMINI
CHIOMC	Diptera	Chironomidae	Microtendipes	CHIRONOMINI
CHIOMI	Diptera	Chironomidae	Microchironomus	CHIRONOMINI

CHIOPA	Diptera	Chironomidae	Paracladopelma	CHIRONOMINI
CHIOPC	Diptera	Chironomidae	Parachironomus	CHIRONOMINI
CHIOPH	Diptera	Chironomidae	Phaenospectra	CHIRONOMINI
CHIOPO	Diptera	Chironomidae	Polypedilum	
CHIOPR	Diptera	Chironomidae	Paralauterbornie	CHIRONOMINI
CHIOPT	Diptera	Chironomidae	Paratendipes	CHIRONOMINI
CHIOSE	Diptera	Chironomidae	Stenochironomus	CHIRONOMINI
CHIOSI	Diptera	Chironomidae	Stichtochironomu	CHIRONOMINI
CHIOSR	Diptera	Chironomidae	Sergentia	CHIROMINI
CHIOST	Diptera	Chironomidae	Stelechomyia	CHIRONOMINI
CHIR	Diptera	Chironomidae		
CHIRAB	Diptera	Chironomidae	Ablabesmyia	
CHIRBR	Diptera	Chironomidae	Brillia	
CHIRBU	Diptera	Chironomidae	Brundiniella	
CHIRCL	Diptera	Chironomidae	Cladotanytarsus	
CHIRCO	Diptera	Chironomidae	Corynoneura	
CHIRCR	Diptera	Chironomidae	Cryptochironomus	
CHIRDI	Diptera	Chironomidae	Diamesa	
CHIRDP	Diptera	Chironomidae	Diplocladius	
CHIREI	Diptera	Chironomidae	Einfeldia	
CHIRER	Diptera	Chironomidae	Euryhapsis	
CHIREU	Diptera	Chironomidae	Eukiefferiella	
CHIREUC	Diptera	Chironomidae	Eukiefferiella	coerulesc
CHIRHE	Diptera	Chironomidae	Helopelopia	
CHIRHY	Diptera	Chironomidae	Hydrobaenus	
CHIRLA	Diptera	Chironomidae	Labrundinia	
CHIRLR	Diptera	Chironomidae	Larsia	
CHIRMA	Diptera	Chironomidae	Macropelopia	
CHIRMI	Diptera	Chironomidae	Micropsectra	
CHIRNA	Diptera	Chironomidae	Nanocladius	
CHIRNI	Diptera	Chironomidae	Nilotanypus	
CHIRNT	Diptera	Chironomidae	Natarsia	
CHIROR	Diptera	Chironomidae	Orthocladius	
CHIRPA	Diptera	Chironomidae	Paracladius	

CHIRPI	Diptera	Chironomidae	Paratrichocladiu	.
CHIRPP	Diptera	Chironomidae	Paraphenocladius	
CHIRPR	Diptera	Chironomidae	Procladius	
CHIRPT	Diptera	Chironomidae	Paratanytarsus	
CHIRRE	Diptera	Chironomidae	Rheotanytarsus	
CHIRRH	Diptera	Chironomidae	Rheocricotopus	
CHIRST	Diptera	Chironomidae	Stempellinella	
CHIRTA	Diptera	Chironomidae	Tanytarsini	
CHIRTH	Diptera	Chironomidae	Thienemanniella	
CHIRTI	Diptera	Chironomidae	Thienemannimyia	
CHIRTN	Diptera	Chironomidae	Tanytarsus	
CHIRTV	Diptera	Chironomidae	Tvetenia	
CHIRTY	Diptera	Chironomidae	Tanypus	
COENAR	Odonata	Coenagrionidae	Argia	
COENARP	Odonata	Coenagrionidae	Argia	plana
COENCO	Odonata	Coenagrionidae	Coenagrion	
COENEN	Odonata	Coenagrionidae	Enallagma	
COL	Collembola			
CORBCO	Pelecypoda	Corbiculidae	Corbicula	
CORDSOE	Odonata	Corduliidae	Somatochlora	ensigera
CORI	Hemiptera	Corixidae		
CORISI	Hemiptera	Corixidae	Sigara	
CORYCOC	Megaloptera	Corydalidae	Corydalus	cornutus
COSMPY	Lepidoptera	Cosmopterigidae	Pyroderces	
CRANSYB	Amphipoda	Crangonyctidae	Synurella	bifurca
CULI	Diptera	Culicidae		
CULICU	Diptera	Culicidae	Culicoides	
CURC	Coleoptera	Curculionidae		
DEC	Decapoda			
DIXIDI	Diptera	Dixidae	Dixa	
DOLI	Diptera	Dolichopodidae		
DOLICL	Diptera	Dolichopodidae	Clinocera	
DRYOHEB	Coleoptera	Dryopidae	Helichus	basalis
DRYOHEL	Coleoptera	Dryopidae	Helichus	lithophilus

DRYOHES	Coleoptera	Dryopidae	Helichus	striatus
DYTI	Coleoptera	Dytiscidae		
DYTIAG	Coleoptera	Dytiscidae	Agabus	
DYTIAI	Coleoptera	Dytiscidae	Agibinus	
DYTICO	Coleoptera	Dytiscidae	Copelatus	
DYTIHD	Coleoptera	Dytiscidae	Hydroporus	
DYTIHY	Coleoptera	Dytiscidae	Hydaticus	
ELMICLO	Coleoptera	Elmidae	Cleptelmis	ornata
ELMIDU	Coleoptera	Elmidae	Dubiraphia	
ELMIDUB	Coleoptera	Elmidae	Dubiraphia	bivattata
ELMIHEC	Coleoptera	Elmidae	Heterlimnius	corpulentus
ELMIMAG	Coleoptera	Elmidae	Macronychus	glaboratus
ELMIMI	Coleoptera	Elmidae	Microcylloepus	
ELMIOP	Coleoptera	Elmidae	Optioservus	
ELMIOPI	Coleoptera	Elmidae	Optioservus	immunis
ELMIOPO	Coleoptera	Elmidae	Optioservus	ozarkensis
ELMIST	Coleoptera	Elmidae	Stenelmis	
EMPICH	Diptera	Empididae	Chelifera	
EMPIEM	Diptera	Empididae	Empididae	
EMPIHE	Diptera	Empididae	Hemerodromia	
EPHE	Ephemeroptera	Ephemeridae		
EPHEEAU	Ephemeroptera	Ephemerellidae	Eurylophella	aestiva
EPHIHEL	Ephemeroptera	Ephemeridae	Hexagenia	limbata
EPHY	Diptera	Ephydridae		
ERPOERP	Hirudinea	Erpobdellidae	Erpobdella	punctata
ERPOMOM	Hirudinea	Erpobdellidae	Mooreobdella	microstoma
GAMMGA	Amphipoda	Gammaridae	Gammarus	
GAS	Gastropoda			
GELAGE	Hemiptera	Gelastocoridae	Gelastocoris	
GERRAQ	Hemiptera	Gerridae	Aquarius	
GERRGER	Hemiptera	Gerridae	Gerris	remigis
GERRME	Hemiptera	Gerridae	Metrobates	
GERRRH	Hemiptera	Gerridae	Rheumatobates	
GERRTR	Hemiptera	Gerridae	Trepobates	

GLOOAG	Trichoptera	Glossosomatidae	Agapetus	
GLOOAGI	Trichoptera	Glossosomatidae	Agapetus	illini
GLOSGLC	Hirudinea	Glossiponiidae	Glossophonia	complanata
GLOSHET	Hirudinea	Glossiponiidae	Helobdella	triserialis
GOMPOPS	Odonata	Gomphidae	Ophiogomophus	severus
GOMPSTA	Odonata	Gomphidae	Stylogomphus	albistostyl
GOMPSTY	Odonata	Gomphidae	Stylogomphus	albistylus
GYRI	Coleoptera	Gyrinidae		
GYRIDI	Coleoptera	Gyrinidae	Dineutus	
HALIHA	Coleoptera	Haliplidae	Haliplus	
HALIPE	Coleoptera	Haliplidae	Peltodytes	
HEBRLI	Hemiptera	Hebridae	Lipogomphus	
HELIHE	Trichoptera	Helicopsychidae	Helicopsyche	
HELIHEB	Trichoptera	Helicopsychidae	Helicopsyche	borealis
HELOHE	Coleoptera	Helophoridae	Helophorus	
HEPT	Ephemeroptera	Heptageniidae	Heptageniidae	
HEPTCI	Ephemeroptera	Heptageniidae	Cinygmula	
HEPTHE	Ephemeroptera	Heptageniidae	Heptagenia	
HEPTHED	Ephemeroptera	Heptageniidae	Heptagenia	diabasa
HEPTHEF	Ephemeroptera	Heptageniidae	Heptagenia	flavescens
HEPTLE	Ephemeroptera	Heptageniidae	Leucrocuta	
HEPTNI	Ephemeroptera	Heptageniidae	Nixe	
HEPTSE	Ephemeroptera	Heptageniidae	Stenonema	
HEPTSEF	Ephemeroptera	Heptageniidae	Stenonema	femoratum
HEPTST	Ephemeroptera	Heptageniidae	Stenacron	
HEPTSTI	Ephemeroptera	Heptageniidae	Stenacron	interpunctatum
HETEHE	Coleoptera	Heteroceridae	Heterocerus	
HIR	Hirudinea			
HYDCHY	Coleoptera	Hydrochidae	Hydrochus	
HYDMHY	Hemiptera	Hydrometridae	Hydrometra	
HYDP	Coleoptera	Hydrophilidae		
HYDPBE	Coleoptera	Hydrophilidae	Berosus	
HYDPENH	Coleoptera	Hydrophilidae	Enochrus	hamiltoni
HYDPHYF	Coleoptera	Hydrophilidae	Hydrobius	fuscipes

HYDPPA	Coleoptera	Hydrophilidae	Paracymus	
HYDPTR	Coleoptera	Hydrophilidae	Tropisternus	
HYDPTRL	Coleoptera	Hydrophilidae	Tropisternus	lateralis
HYDR	Gastropoda	Hydrobiidae		
HYDRAC	Hydracarina	Hydrachnidae	Acarina	
HYDS	Trichoptera	Hydropsychidae	Hydropsychidae	
HYDSCE	Tricoptera	Hydropsychidae	Ceratopsyche	
HYDSCH	Trichoptera	Hydropsychidae	Cheumatopsyche	
HYDSHY	Trichoptera	Hydropsychidae	Hydropsyche	
HYDT	Trichoptera	Hydroptilidae		
HYDTHY	Trichoptera	Hydroptilidae	Hydroptila	
HYDTOC	Trichoptera	Hydroptilidae	Ochrotrichia	
HYDTOX	Trichoptera	Hydroptilidae	Oxyethira	
HYGRAT	Hydracarina	Hygrobatidae	Atractides	
HYGRHY	Hydracarina	Hygrobatidae	Hygrobates	
ISO	Isopoda			
ISOTFO	Collembola	Isotomidae	Folsomia	
LEBELE	Hydracarina	Lebertiidae	Lebertia	
LEPIAR	Lepidoptera	Noctuidae	Archanara	
LEPO	Ephemeroptera	Leptophlebiidae		
LEPOHAA	Ephemeroptera	Leptophlebiidae	Habrophlebiodes	american
LEPOLE	Ephemeroptera	Leptophlebiidae	Leptophlebia	
LEPOLEN	Ephemeroptera	Leptophlebiidae	Leptophlebia	nebulosa
LEPT	Trichoptera	Leptoceridae		
LEPTNE	Trichoptera	Leptoceridae	Nectopsyche	
LEPTPA	Ephemeroptera	Leptophlebiidae	Paraleptophlebia	
LEPTTR	Trichoptera	Leptoceridae	Trianodes	
LESTLE	Odonata	Lestidae	Lestes	
LEUCLE	Plecoptera	Leuctridae	Leuctra	
LEUCLET	Plecoptera	Leuctridae	Leuctra	tenuis
LEUCZE	Plecoptera	Leuctridae	Zealeuctra	
LEUCZEC	Plecoptera	Leuctridae	Zealeuctra	claasseni
LIBE	Odonata	Libellulidae		
LIMELI	Hydracarina	Limnesiidae	Limnesia	

LIMN	Trichoptera	Limnephilidae		
LIMNANB	Trichoptera	Limnephilidae	Anabolia	bimaculata
LIMNLID	Trichoptera	Limnephilidae	Limnephilus	diversus
LIMNNE	Trichoptera	Limnephilidae	Neophylax	
LIMNNEF	Trichoptera	Limnephilidae	Neophylax	fuscus
LIMNPY	Trichoptera	Limnephilidae	Pycnopsyche	
LUMB	Oligochaeta	Lumbricidae	Lumbricidae	
LYMNLY	Gastropoda	Lymnaeidae	Lymnaea	
MUSC	Diptera	Muscidae		
MUSCMU	Diptera	Muscidae	Muscidae	
NEA	Nematomorpha			
NECTNE	Lepidoptera	Nectuidae	Nectuidae	
NEM	Nematoda			
NEMOAM	Plecoptera	Nemouridae	Amphinemura	
NEMOAMD	Plecoptera	Nemouridae	Amphinemura	delosa
NEPIRAF	Hemiptera	Nepidae	Rantra	fusca
OLI	Oligochaeta			
OLIGIS	Ephemeroptera	Oligoneuriidae	Isonychia	
OLIGISR	Ephemeroptera	Oligoneuriidae	Isonychia	rufa
PERLAC	Plecoptera	Perlidae	Acroneuria	
PERLACF	Plecoptera	Perlidae	Acroneuria	frisoni
PERLAG	Plecoptera	Perlidae	Agnetina	
PERLAGC	Plecoptera	Perlidae	Agnetina	capitata
PERLAGF	Plecoptera	Perlidae	Agnetina	flavescens
PERLATR	Plecoptera	Perlidae	Attaneuria	ruralis
PERLPE	Plecoptera	Perlidae	Perlesta	
PERLPEC	Plecoptera	Perlidae	Perlesta	cinctipes
PERLPED	Plecoptera	Perlidae	Perlesta	decipiens
PERLPR	Plecoptera	Perlidae	Perlinella	
PEROCLC	Plecoptera	Perlodidae	Clioperla	clio
PEROCU	Plecoptera	Perlodidae	Cultus	
PEROIS	Plecoptera	Perlodidae	Isoperla	
PEROPE	Plecoptera	Perlodidae	Perlodidae	
PHILCH	Trichoptera	Philopotamidae	Chimarra	

PHILCHA	Trichoptera	Philopotamidae	Chimarra	atterrima
PHRYPTS	Trichoptera	Phryganeidae	Ptilostomis	semifasciata
PHYS	Gastropoda	Physidae		
PHYSPH	Gastropoda	Physidae	Physa	
PLAN	Tricladida	Planariidae		
PLANDU	Tricladida	Planariidae	Dugesia	
PLANDUT	Tricladida	Planariidae	Dugesia	tigrina
PLAO	Gastropoda	Planorbidae		
PLAOGY	Gastropoda	Planorbidae	Gyraulus	
PLAOPH	Gastropoda	Planorbidae	Physella	
PLAOPR	Gastropoda	Planorbidae	Promentus	
PLE	Plecoptera			
PLEU	Gastropoda	Pleuroceridae		
PLEUPL	Gastropoda	Pleuroceridae	Pleurocera	
POLCE	Trichoptera	Polycentropodidae	Cernotina	
POLYPO	Trichoptera	Polycentropodidae	Polycentropus	
POLYPOC	Trichoptera	Polycentropodidae	Polycentropus	cinerus
PSEPEC	Coleoptera	Psephenidae	Ectopria	
PSEPPS	Coleoptera	Psephenidae	Psephenus	
PSEPPSH	Coleoptera	Psephenidae	Psephenus	herricki
PSYC	Diptera	Psychodidae		
PSYCPS	Diptera	Psychodidae	Psychoda	
PSYHLYD	Trichoptera	Psychomyiidae	Lype	diversa
PSYHPS	Trichoptera	Psychomiidae	Psychomyia	
PYRAPE	Lepidoptera	Pyralidae	Petrophila	
SALD	Hemiptera	Saldidae		
SALIPE	Hemiptera	Salididae	Pentacora	
SCIR	Coleoptera	Scirtidae		
SCIRSC	Coleoptera	Scirtidae	Scirtes	
SIALSI	Megaloptera	Sialidae	Sialis	
SIALSIV	Megaloptera	Sialidae	Sialis	velta
SIMUSI	Diptera	Simuliidae	Simulium	
SISYSIV	Neuroptera	Sisyridae	Sisyra	vicaria
SPERSE	Hydracarina	Sperchonidae	Sperchonopsis	

SPERSP	Hydracarina	Sperchonidae	Sperchon	
SPHA	Pelecypoda	Sphaeriidae		
SPHASP	Pelecypoda	Sphaeriidae	Sphaerium	
STAPBL	Coleoptera	Staphylinidae	Bledius	
STAPCA	Coleoptera	Staphylinidae	Carpelimus	
STAPST	Coleoptera	Staphylinidae	Stenus	
STRA	Diptera	Stratiomyidae		
STRACA	Diptera	Stratomyidae	Caloparyphus	
SYRP	Diptera	Syrphidae		
TABA	Diptera	Tabanidae		
TABACH	Diptera	Tabanidae	Chrysops	
TABATA	Diptera	Tabanidae	Tabanus	
TAENTA	Plecoptera	Taeniopterygidae	Taeniopteryx	
TAENTAB	Plecoptera	Taeniopterygidae	Taeniopteryx	burksi
TALIHYA	Amphipoda	Talitridae	Hyalella	azteca
TANYTA	Diptera	Tanypodinae	Tanypodinae	
TENICR	Diptera	Tenipedidae	Cricotopus	
TENICRB	Diptera	Tenipedidae	Cricotopus	bicinctus
TENICRC	Diptera	Tenipedidae	Cricotopus	cylindraceus
TENICRT	Diptera	Tenipedidae	Cricotopus	tremulus
TETR	Orthoptera	Tetrigidae		
THIAEL	Gastropoda	Thiaridae	Elimia	
TIPUAN	Diptera	Tipulidae	Antocha	
TIPUDI	Diptera	Tipulidae	Dicranota	
TIPUHE	Diptera	Tipulidae	Hexatoma	
TIPULI	Diptera	Tipulidae	Limonia	
TIPUPS	Diptera	Tipulidae	Pseudolimnophila	
TIPUTI	Diptera	Tipulidae	Tipula	
TORTBA	Lepidoptera	Tortricidae	Bactra	
TRICTR	Ephemeroptera	Tricorythidae	Tricorythodes	
TUBIILM	Oligochaeta	Tubificidae	Ilyodrilus	mastix
TUR	Turbellaria			
UNK	unknown			
VELIMI	Hemiptera	Veliidae	Microvelia	

VELIMIB	Hemiptera	Veliidae	Microvelia	buenoi
VELIPA	Hemiptera	Veliidae	Paravelia	
VELIRHO	Hemiptera	Veliidae	Rhagovelia	obesa

Appendix F. Tolerance Values for Families of Stream Arthropods in the Four Prairie Streams. Values from Hilsenhoff (1988).

Order	Family	Tolerance Value
Amphipoda	Crangonyctidae	8
Amphipoda	Gammaridae	4
Amphipoda	Talitridae	8
Coleoptera	Dryopidae	5
Coleoptera	Elmidae	4
Coleoptera	Psephenidae	4
Diptera	Athericidae	2
Diptera	Ceratopogonidae	6
Diptera	Chironomidae (other)	6
Diptera	Chironomidae (red-blooded)	8
Diptera	Culicidae	6
Diptera	Dixidae	6
Diptera	Empididae	6
Diptera	Ephydridae	6
Diptera	Psychodidae	10
Diptera	Simuliidae	6
Diptera	Stratiomyidae	6
Diptera	Tabanidae	6
Diptera	Tenipedidae	6
Diptera	Tipulidae	3
Ephemeroptera	Ameletidae	4
Ephemeroptera	Baetidae	4
Ephemeroptera	Caenidae	7
Ephemeroptera	Ephemerellidae	1
Ephemeroptera	Ephemeridae	4
Ephemeroptera	Heptageniidae	4
Ephemeroptera	Leptophlebiidae	2
Ephemeroptera	Oligoneuriidae	2
Ephemeroptera	Tricorythidae	4
Isopoda	-	8
Isopoda	Asellidae	8
Lepidoptera	Pyralidae	5
Lepidoptera	Tortricidae	5
Megaloptera	Chauliodinae	4
Megaloptera	Corydalidae	0
Megaloptera	Sialidae	4
Odonata	Aeshnidae	3

Appendix F. (continued)

Order	Family	Tolerance Value
Odonata	Calopterygidae	5
Odonata	Coenagrionidae	5
Odonata	Corduliidae	5
Odonata	Gomphidae	1
Odonata	Lestidae	9
Odonata	Libellulidae	9
Plecoptera	Leuctridae	0
Plecoptera	Nemouridae	2
Plecoptera	Perlidae	1
Plecoptera	Perlodidae	2
Plecoptera	Taeniopterygidae	2
Trichoptera	Glossosomatidae	0
Trichoptera	Helicopsychidae	3
Trichoptera	Hydropsychidae	4
Trichoptera	Hydroptilidae	4
Trichoptera	Leptoceridae	4
Trichoptera	Limnephilidae	4
Trichoptera	Philopotamidae	3
Trichoptera	Phryganeidae	4
Trichoptera	Polycentropodidae	6
Trichoptera	Psychomyiidae	2

Appendix G. The Macroinvertebrate Biomonitoring Status Report for Pipestone National Monument, 1997.

Prairie Cluster Long-Term

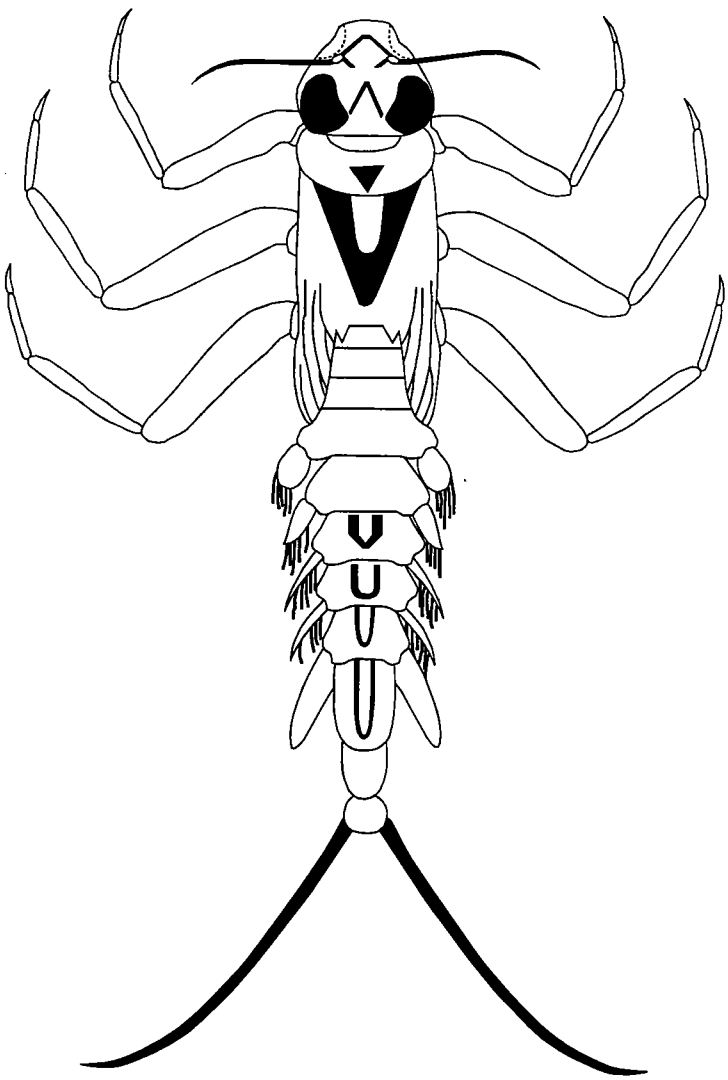
Ecological Monitoring Program

Program Report

97-001

Annual Status Report:

1997 Stream Macroinvertebrate Biomonitoring for Pipestone National Monument



Annual Status Report:

1997 Stream Macroinvertebrate Biomonitoring for Pipestone National Monument

by

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February 5, 1999

1.0 INTRODUCTION

The structure of aquatic macroinvertebrate communities is controlled by the physical and chemical characteristics of a stream, particularly water quality and habitat structure. Consequently, changes in these variables can alter aquatic macroinvertebrate community structure. Since aquatic macroinvertebrates have relatively long lives they integrate the high variability of chemical and physical factors over their lifespan, making them ideal for monitoring stream water quality (Rosenberg and Resh 1993). At Pipestone National Monument (PIPE), Minnesota, the National Park Service began macroinvertebrate biomonitoring in Pipestone Creek in 1989 (Boyle et al. 1990). This dataset forms the baseline against which the 1997 monitoring data is compared.

2.0 OBJECTIVES

The objective of macroinvertebrate biomonitoring at PIPE is to determine the annual status of macroinvertebrate community structure to estimate stream water quality in comparison to the baseline dataset, using four metrics of community structure. As monitoring continues, the resulting long-term datasets can be used to estimate trends over time.

3.0 METHODS

3.1 Field and Laboratory Procedures

Macroinvertebrates were collected from riffles at two sites in Pipestone Creek (PPS1 and PPS2) with a square-foot (0.0929 m^2) Surber sampler having a capture net mesh size of 1000 µm. The sampler was placed in riffles with sufficient current and depth to wash dislodged organisms into the capture net. The substrate within the sampler was disturbed with a hand rake to a depth of 8-10 cm to dislodge organisms. Individual rocks within the sampler were also scrubbed to dislodge clinging organisms. In 1997, five replicate samples were collected from different riffles on each sampling date (June 26, August 13, and either September 17 (PPS2) or 23 (PPS1)). Samples were preserved in 5% formalin. The samples

were picked, sorted, identified to the required taxonomic level, and enumerated by technicians at the Missouri Cooperative Fish and Wildlife Research Unit, University of Missouri - Columbia. Sampling and identification procedures are described in detail in Peterson et al. (1999).

3.2 Community Structure Metrics

Changes in macroinvertebrate community structure can occur from habitat alterations as well as changes in water quality. Since different measures of community structure also differ in their sensitivity to habitat vs. water quality changes (Figure 1), at least two of the community structure metrics must change significantly before concluding that significant changes in community structure have occurred. In addition, the habitat assessment data must not suggest physical alterations as the cause of significant changes before concluding that community changes are due to altered water quality. However, the selected metrics are less sensitive to habitat alterations than to changes in water quality, so habitat changes would have to be substantial to be implicated as the cause of a significant finding (Rabeni et al. 1997).

The four community structure metrics used at PIPE are:

1. Family biotic index (FBI). This index uses family-specific pollution tolerance values to calculate index scores for each replicate sample. It is calculated as: $FBI = (\Sigma n_i a_i/N)-0.18$; where N is the total number of individuals in the sample, and n_i is the total number of individuals in the *i*th family, and a_i is the tolerance value for the *i*th family. FBI values are related to water quality as follows (Hilsenhoff 1988):

Excellent	0.00 - 3.50
Very good	3.51 - 4.50
Good	4.51 - 5.50
Fair	5.51 - 6.50
Fairly poor	6.51 - 7.50
Poor	7.51 - 8.50
Very poor	8.51 - 10.00

2. Ratio of Ephemeroptera, Trichoptera, and Plecoptera (EPT) abundance to abundance of EPT + Chironomidae. In general, EPT taxa are pollution intolerant, whereas Chironomidae are pollution tolerant. Thus, larger values indicate better water quality (Resh and Grodhaus 1983). The EPT ratio (R) is calculated for each replicate sample as: R = EPT/[EPT + C]; where EPT is the abundance of EPT taxa, and C is the abundance of Chironomidae.

3. Family richness (total number of families). Low family richness can indicate a stream affected by one or more environmental stressors (Resh and Grodhaus 1983). Family richness was calculated for each replicate sample.

4. Family diversity. Diversity metrics indicate how the total density of individuals is distributed among the families of a macroinvertebrate community. Maximum diversity occurs when the individuals are distributed as evenly as possible among families and indicates better stream quality (Resh and Jackson 1993). The Shannon-Wiener diversity index (H′) was calculated for each replicate as: $H' = -\Sigma(n_i/N)\ln(n_i/N)$; where N is the total number of individuals in the sample, and n_i is the total number of individuals in the *i*th family.

3.3 Data Analysis

To evaluate the status of the macroinvertebrate community in 1997, the metrics were calculated as above and compared by one-way analysis of variance (ANOVA) to the means calculated for the June 1989 baseline dataset. Prior to carrying out these ANOVA's, the data were tested for the assumptions of normality and homogeneity of variances. The diversity data were not normally distributed and were log transformed prior to the analysis.

4.0 RESULTS and DISCUSSION

The FBI index did not change significantly at either site (Tables 1 and 2), although the water quality rating fell from'very good' to 'good' at PPS1. The FBI water quality rating at PPS2 remained 'very good.' Similary, family richness showed no change at PPS1, but declined significantly at PPS2. Family diversity values declined significantly between years at both sites. Thus, the latter two metrics show that the number of taxa at PPS1 was unchanged

although the evenness of the distribution of individuals among taxa declined. At PPS2 both the number of taxa and the distribution of individuals among taxa both decreased. The EPT ratio declined in PPS1, indicating poorer water quality, but was not significantly different between years at PPS2. Thus, the net assessment for the sites at PIPE is that a decline in water quality has occurred at both sites.

The 1997 habitat assessments show only very slight changes in temperature, gauge height, depth (Figure 2) or sediment grain size distribution (Figure 3) over the summer. Current velocity, however, was substantially greater in June than in the other two months, and could possible have influenced the 1997 metrics. However, interpretation of the habitat data is problematical since no baseline habitat data exist.

Figures 4-7 show the changes in community metrics at the PIPE sites since biomonitoring began. Also shown for all metrics are values from other studies. Within the last three years when sampling began to consistently follow the draft biomonitoring protocol (Peterson et al. 1999), all metrics except family diversity have been relatively stable, or shown some improvement. Family diversity values have continued to decline at both sites over the past three years.

Relative to other studies, the recent FBI data show that PIPE data falls toward the mid-range of values (average water quality) for the streams used by Hilsenhoff (1988) in his classification (Figure 4). The minimum richness value given by Rabeni et al. (1997) for 25 Ozark streams was greater than all values from PIPE sites except PPS2 in 1992 (Figure 5), indicating a substantial difference from other streams in the region. However, diversity values for PIPE streams are generally higher than the minimum value reported by Rabeni et al. (1997) for 16 Missouri prairie streams (Figure 6). The EPT ratios are also greater than the minimum reported by Rabeni et al. (1997), but are less than half the mean value they report (Figure 7).

Although direct comparison of the PIPE data with the study by Rabeni et al. (1997) suggest poorer water quality at PIPE than other prairie streams, the comparisons should be made with caution. The other studies encompass more seasonal, geographic, and habitat variability than the PIPE dataset. Also, Rabeni et al. (1997) were primarily interested in establishing baseline conditions for reference streams, i.e. relatively pristine systems. Thus, many other streams probably have water quality similar to the streams at PIPE. However,

reference datasets like Rabeni et al. (1997) are valuable in showing how different PIPE water quality is compared to relatively pristine systems.

Changes of the magnitude shown for most of the metrics in Tables 1-3 can probably be readily detected by any metric. Future changes may well be more subtle, and could be missed since the baseline dataset comprises only a single June sample. A single sample will not encompass the environmental variability of an entire season (Peterson 1997; Rabeni et al. 1997), and thus cannot be strictly comparable to larger datasets encompassing the entire season. It would be wise to establish a new baseline using the earliest year in which sampling conformed to the draft macroinvertebrate monitoring protocol (Peterson et al. 1999), or to include such a year in the analyses along with the 1989 dataset. In addition, the Surber capture net should be changed to a mesh size of ca. 200 μm. Larger mesh sizes miss many smaller taxa, especially chironomids, and thus may bias the outcome of many of the metrics. The field notes from the 1997 sampling season describe losing macroinvertebrates through the large mesh capture net.

5.0 REFERENCES

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Table 1. Assessment of the status of water quality in Pipestone Creek (site PPS1) in 1997. Results are: N = No change, D = Deleterious impact, and I = Improvement over the 1989 baseline year.

Date	FBI	EPT Ratio	Family Richness	Family Diversity
June 26				
Replicate 1	1.58	0.40	11	1.54
Replicate 2	5.85	0.03	12	0.29
Replicate 3	5.18	0.22	10	1.24
Replicate 4	4.69	0.13	11	0.61
Replicate 5	5.80	0.35	20	1.66
August 13				
Replicate 1	6.49	0.00	3	0.41
Replicate 2	4.82	0.50	1	0.00
Replicate 3	3.82	1.00	1	0.00
Replicate 4	3.82	0.32	1	0.00
Replicate 5	4.49	0.43	3	1.10
September 23				
Replicate 1	4.93	0.13	15	1.28
Replicate 2	5.82	0.00	1	0.00
Replicate 3	4.93	0.44	2	0.69
Replicate 4	5.82	0.00	1	0.00
Replicate 5	4.93	0.63	5	1.21
Mean	4.86	0.31	6.5	0.67
1989 mean	3.99	0.70	10.6	1.78
F_{PROB}	0.1540	0.0069	0.1615	0.0219
Result	N	D	N	D

Net Assessment: Decline in water quality

Table 2. Assessment of the status of water quality in Pipestone Creek (site PPS2) in 1997. Results are: N = No change, D = Deleterious impact, and I = Improvement over the 1989 baseline year.

Date	FBI	EPT Ratio	Family Richness	Family Diversity
June 26				
Replicate 1	3.43	0.62	10	1.91
Replicate 2	3.12	0.22	7	1.48
Replicate 3	2.66	0.66	7	1.44
Replicate 4	5.82	0.14	3	0.41
Replicate 5	2.90	0.65	6	1.23
August 13				
Replicate 1	5.15	0.00	4	0.89
Replicate 2	5.32	0.00	3	0.56
Replicate 3	4.15	1.00	4	1.24
Replicate 4	4.93	0.32	8	1.10
Replicate 5	4.39	0.59	6	1.10
September 17				
Replicate 1	2.56	0.17	5	1.12
Replicate 2	3.53	1.00	2	0.41
Replicate 3	5.42	0.00	2	0.50
Replicate 4	3.53	0.11	5	1.32
Replicate 5	5.90	0.00	4	0.33
Mean	4.19	0.37	5.1	1.00
1989 mean	4.34	0.59	11.0	1.74
F_{PROB}	0.7937	0.1902	0.0001	0.0158
Result	N	N	D	D

Net Assessment: Decline in water quality